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SAMPLING STRATEGIES FOR ESTIMATING MOTH SPECIES DIVERSITY USING A LIGHT TRAP IN A NORTHEASTERN SOFTWOOD FOREST

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ABSTRACT. A 22-watt black-light trap was operated for 29 nights within a forest canopy in the Maritime Lowlands Ecoregion of the Acadian Forest. The species-abundance frequency distribution (pattern of species abundance) was a good fit to the log series model and this model was used for subsequent data analysis. No single-night sample adequately estimated the log series *alpha* index of diversity based on the total catch; some sampling effort was required each night. Each night's catch was separated into 16, 30-minute samples. The *alpha* index of diversity for the summed catch for each time-period was compared with the overall *alpha* based on the total catch. A strategy that involved operating the trap for just a 1-hour period each night had no effect on the pattern of species abundance and gave a value for *alpha* equal to that obtained by operating the trap for an 8-hour period each night. This strategy reduced the catch from 6088 to 971 moths and the number of species from 255 to 161. Processing costs associated with the larger sample and any possible negative effect on the moth population caused by removal trapping were greatly reduced. This new sampling strategy is thus useful for comparing indices of species diversity between several sites when data are collected simultaneously, but is of limited use for species-inventory studies.

Additional key words: species-abundance distribution, 30-minute samples, log-series model, partial-night sampling.

In recent years, the challenge to maintain biodiversity on this planet has become a major public concern. Most attention focuses on Neotropical ecosystems (Mares 1992). However, the importance of maintaining Canada's biodiversity was addressed in Environment Canada's Green Plan (Hyslop & Brunton 1991), and the launching, in 1991, of "Canadian Biodiversity" produced by the Canadian Centre of Biodiversity at the Canadian Museum of Nature lends credence to the recent national interest in biodiversity. The values of maintaining current biodiversity have been stated by many authors, and summarized by Ehrlich (1990) into ethical, aesthetic, economic, and 'ecosystem services.' Salwasser (1990) added the legal obligation for conserving biological diversity. Intimately linked with the concept of maintaining biodiversity, and especially protection of areas rich in species, is the need for a "quick and dirty survey to chart biodiversity of the planet" (Roberts 1988), a view reiterated by Ehrlich (1992). The 'quick and dirty' approach does not advocate poor science; rather, it recognizes that the scope of diversity from individual gene systems through populations of species, communities, ecosystems, and ultimately all life in the biosphere (Wilson 1988) cannot be addressed in the short-term. It suggests that studies should be focused on certain taxonomic groups over an extensive area. The hope is that areas with many species or high endemism in the selected groups will reflect similarly high values for other groups (Roberts 1988). Because of logistic and knowledge constraints, the number of species within a community can be determined for only a limited number of taxonomic groups.

This study addresses just one segment of biodiversity, i.e., the diversity of moths in a single ecosystem. Diversity is used here to mean the number of species and their relative abundance (Magurran 1988), and to prevent ambiguity we will always use 'species diversity' where appropriate. Relative abundance is considered in the form of species-abundance frequency distributions, which show the relationship between the abundance of individuals and the number of species possessing that abundance (May 1975); abbreviated in this paper as the pattern of species abundance. The ecosystem studied is one locality in the Maritime Lowlands Ecoregion of the Acadian Forest (Loucks 1962).

The use of the moth community, in the 15 families used in this study (see Appendix), as an exemplar of the species diversity of this ecosystem has advantages that include the relative ease of identification at the species level, the somewhat standardized sampling methodology (Williams 1951, Williams et al. 1955, Taylor & French 1974, Bowden 1982), and the high correlation of insects, in general, with the spatial, architectural, and taxonomic diversity of plants (Southwood et al. 1979).

No community consists of species of equal abundance (Magurran 1988). It is normally the case that the majority of species are rare while a number are moderately common with the remaining few species being very abundant (Williams 1964, May 1975, Pielou 1975, Southwood 1978, Magurran 1988). Within this general distributional form, communities have characteristically different patterns of species abundance which remain stable despite changes in species composition (Pielou 1975, May 1976, Kempton 1979). The pattern of species abundances at a site allows for comparison with similar sites that have different mixes of species, and a change in the pattern of abundance at one site has been shown to be a useful indicator of environmental disturbance (Kempton & Taylor 1974, Taylor et al. 1978, Kempton 1979).

Four main species-abundance models (the geometric series, the logarithmic series, the log normal distribution, and MacArthur's broken stick model) have been developed to describe species diversity in terms of an 'index parameter' as well as the pattern of species abundance. In addition there are several non-parametric indices based on the proportional abundances of species (May 1975, Southwood 1978, Magurran 1988).

The log series model was the first to describe the pattern of species abundance (Fisher 1943). Since then it has been found to have a wide application for catches of many invertebrates, e.g., moths in light traps (Williams 1943, 1945, 1964, Taylor & Brown 1972, Taylor & French 1974, Kempton & Taylor 1974, Taylor et al. 1976, 1978, Taylor 1986), Ichneumonidae (Owen & Chanter 1970), cockroaches (Wolda 1983), Psocoptera (Broadhead & Wolda 1985), Hymenoptera (Noyes 1989), and the community of phytophagous arthropods on apple (Brown & Adler 1989). Its wide applicability is because it is based on the abundances of the species with medium abundance rather than the very abundant and very rare species (Taylor et al. 1976, Kempton 1979, Brown & Adler 1989).

The log series is a simple two-parameter model, with two defining multispecies population parameters, *chi* and *alpha*. *Chi* is devoted to sample characteristics and varies with sample size as it is a function of the mean number of individuals per species. *Alpha* is independent of sample size and characterizes the required population quality (Kempton & Taylor 1974). Fisher's (1943) initial suggestion was that *alpha* might be useful as a measure of 'species richness' when comparing samples. Williams (1943) suggested that the parameter *alpha* be known as a community's 'index of diversity.' Later he recognized that this term was applicable to other functions having the same properties and referred to Fisher's *alpha* as 'diversity calculated on the basis of the logarithmic series' (Williams 1964). The log series model can be derived from two statistics, *S*, the total number of species, and *N*, the total number of moths. It is a discontinuous frequency series with an infinite number of terms:

$$n_1, n_1\chi/2, n_1\chi^2/3, n_1\chi^3/4, \ldots,$$

where n_1 is the number of species with 1 individual and successive terms with 2, 3, 4, etc. individuals, and χ (*chi*) is a constant <1 (Williams 1947).

The log normal model was compared with the log series model by Kempton and Taylor (1974) in an analysis of moth catches from light traps at 18 sites for four successive years in an attempt to quantify intuitively recognized properties of habitats. This comparison found that samples from stable environments were best fitted by the log series whereas those from highly perturbed sites better fitted the log normal. Their overall conclusion was that *alpha* of the log series was the superior diversity discriminant, which they defined as a population parameter that behaves consistently within a stable population and responds to changes within, and to differences between, environments (see also Taylor et al. 1976).

The Simpson-Yule diversity statistic and the Shannon-Weaver information statistic (both non-parametric indices) were compared to the log series *alpha* index of diversity by Taylor et al. (1976) using 10 years of light-trap data at one site. Although the log series model was not the ideal description of the pattern of species abundance, the site's environmental stability was better reflected by *alpha* than by either of the other two statistics.

One constraint with using the moth community as an exemplar of species diversity for an ecosystem is the logistics of sorting, counting, and identifying all the individuals in the sample (Taylor 1979). For example, a one-night catch from one trap in Kenya exceeded 6.7 kg (Taylor et al. 1979); 26,300 moths were captured in one light-trap during a nine-month period at Rothamsted (UK) (Williams 1964); 113,256 moths were taken in one light-trap in one year in Kansas (USA) (Williams 1945); 6088 moths were taken in one trap in one month (this study). Methods for reducing the size of catches were detailed by Taylor and Brown (1972), and for subsampling from large catches by Taylor et al. (1979). The objectives of this study were: (1) to describe the species-abundance frequency distribution and determine the log series *alpha* index of diversity, for moths captured in a light-trap in a within-canopy site of a predominantly balsam fir forest during the flight season of the major forest pest, spruce budworm (*Choristoneura fumiferana* (Clemens) (Tortricidae)), and (2) to develop a sampling strategy that reduced the catch to a minimum without causing significant loss of information, measured as no change in the pattern of species abundance and a reduction in the *alpha* value of 5% or less.

METHODS

Moth collection and identification. Beginning on 21 June 1990 (day 1) and ending on 30 July (day 40), one 22-watt black-light trap (Universal Light Trap, Bioquip Products, California) was operated in the Peter Brook study area of the Acadia Forest Experiment Station near

Fredericton, New Brunswick, Canada. For a variety of reasons, full-night trap data are available for only 29 of the potential 40 nights. Intensive studies on the population dynamics of spruce budworm have been in progress at this site since 1986. The physical characteristics and vegetation of the site have been described (Lethiecq & Regnière 1988). Briefly, the study area is composed of 77% balsam fir, *Abies balsamea* (L.) Miller (Pinaceae), 12% red maple, *Acer rubrum* L. (Aceraceae), and eight other tree species. However, the surrounding area is heterogenous and within a 10-km radius contains mixed forest, lakes, streams, sphagnum bogs, large clear-cuts, and roadsides.

The trap, with the lamp at 6.4 m above the ground, was on a platform, 3 × 1.5 m, on a tower within the closed crowns of balsam fir trees; the otherwise touching branches were trimmed to leave a clearing of 3 × 1.5 m. A blue plastic sheet, 1.8 × 2.4 m, was stretched above the platform at a height of 2.4 m above the lamp. This sheet made direct observation of the light impossible from above, although the reflection of the light off of the foliage of the adjacent trees gave a glow to the immediate area which was obvious from the ground.

The trap was equipped with an automatic time-interval collecting device (King et al. 1965, Smith et al. 1973). Each night's total catch consisted of 16, 30-minute sequential samples, beginning with time-period 1 from 2130–2200 h and ending with time-period 16 from 0500–0530 h. On 21 June, day 1, sunset was at 2120 h and sunrise the following morning at 0536 h; on 30 July, day 29, sunset was at 2058 h and sunrise the following morning at 0606 h. At the latitude of New Brunswick, the sky is noticeably lighter about 30 min before sunrise and remains light for 30 min after sunset.

The moths were killed with 1,1,1 trichloroethane. Moths were stored at −17°C until identified and counted. Most specimens were identified with the aid of the literature and confirmed by consulting the Forest Insect and Disease Survey (FIDS) Reference Collection, Canadian Forest Service, Fredericton, which contains specimens identified by the Biological Resources Division (BRD) of the Centre for Land and Biological Resources Research, Ottawa. Genitalia mounts of specimens were made when identification was uncertain. A further 52 species of geometrids were identified by Klaus Bolte and 81 species of noctuids by Don Lafontaine, both at BRD. All moths in the following families were identified to species and counted: Hepialidae; Sesiidae; Cossidae; Limacodidae; Thyatiridae; Drepanidae; Geometridae, except for *Eupithecia*; Lasiocampidae; Saturniidae; Sphingidae; Notodontidae; Arctiidae; Lymantriidae; and Noctuidae. In addition, all specimens of spruce budworm (Tortricidae) were counted. Moths belonging to other families were not identified or recorded. Publications used for species identification were Forbes (1954), McGuffin (1967, 1972, 1977, 1981), Rockburne and Lafontaine (1976), Ferguson (1978), Morris (1980), McCabe (1980), Covell (1984), Laplante (1985), Lafontaine (1987), and Lafontaine and Poole (1991).

Species-abundance frequency distribution. The numbers of species having abundances of 1, 2, 3, . . ., 724 moths (based on the total catch) were compared with the expected numbers from the log series model (Williams 1947) for goodness-of-fit, using the *chi*-square test (Owen & Chanter 1970, Kempton & Taylor 1974, Taylor et al. 1976, Broadhead & Wolda 1985, Magurran 1988, Noyes 1989, Basset & Kitching 1991). The observed abundances covered a large range, 1–724 moths per species, and because many of these 724 abundance classes were zero (e.g., abundance classes 31 and 36 each had two species, but no species had just 32, 33, 34, or 35 moths and thus classes 32–35 were zeros) the abundance classes were grouped into 10 new abundance classes of approximately equal range on the logarithmic (base 2) scale (Kempton & Taylor 1974, Kempton 1975, Taylor et al. 1976). Because the abundance class having >511 moths had an expected frequency of <1 species, this class was pooled with the preceding class to give an expected frequency of >1 species; resulting in just nine abundance classes. This grouping and pooling of abundance classes (see Table 2) resulted in the data set meeting the requirements for the *chi*-square analysis in that no more than 20% of the classes had an expected frequency of <5 species (1 out of 9 did) and no expected frequency was <1 (Zar 1984).

Index of diversity. For the purpose of this study, the 29-night sample from the trap was taken to be the population being sampled. The log series *alpha* index of diversity was determined after rearranging equations (7) and (8) of Williams (1947) to obtain:

$$[1] \qquad (Sx/-\ln(1-x)) - N(1-x) = 0$$

and solving for x using MathCad (1991), and then solving [2] for *alpha*:

$$[2] \qquad alpha = N(1-x)/x$$

This value based on the single 29-night sample was termed 'the overall *alpha*.'

Strategies to reduce sample size. Three data manipulations were employed to determine a strategy that would reduce the size of the sample and thus reduce processing costs and lessen the possible effect of removal trapping on the moth population.

Single-night samples. The first attempt at a sampling strategy was to determine *alpha* for each night's catch and to compare each value with the overall *alpha*. Such a strategy would certainly reduce sample size, but it was not known how representative such an *alpha* based on one night's catch would be of the overall *alpha* based on the total 29-day catch.

'Replicated' single-time-period samples. As each night's catch consisted of 16 sequential 30-minute samples, there were 16 single-time-period samples, with each sample 'replicated' for 29 nights. The *alpha* index of diversity was calculated for each pooled time period (e.g., all the moths trapped during time period 1 were pooled) and compared with the overall *alpha*. If an index equivalent to the overall index could be estimated from a single 30-minute sample taken each night for 29 nights, significant saving in processing costs would occur, i.e., $1 \times 29 = 29$ samples instead of $16 \times 29 = 464$.

Truncated samples. This strategy was based on the results of the single-time-period analysis. As certain time periods gave low *alpha* values, it was argued that these time periods could be eliminated (thus reducing the number of samples, the number of moths, the processing costs) without significant loss of information. Two sub-strategies were employed. The first, termed **early truncation**, was to discard cumulative sequential time periods from the entire data set beginning with all 29 samples from time period 1, then all 58 samples from time period 1 + time period 2, etc. After 15 truncations only the data set from time period 16 remained. The *alpha* index of diversity was calculated from the data set remaining after each truncation and compared with the overall *alpha* to determine the percentage change. Also after each truncation, the pattern of species abundance was compared with that from the log series model using the deviance *chi*-square values (Kempton & Taylor 1974). The second sub-strategy, termed **late truncation**, was similar to early truncation except that all 29 samples from time period 16 were first discarded, then all 58 samples from time periods 16 + 15, etc. Combining selected data sets that remained after early and late truncation (effectively a **double-ended truncation**) gave several sampling strategies that met the goal of reducing sample size without compromising the value for *alpha* or the pattern of species abundance. The durations for these sampling strategies are shown in Table 1.

RESULTS

Totals of 6088 individual moths representing 255 macrolepidoptera species in 15 families were identified from the 29-night catch (see Appendix).

Species-abundance distribution and index of diversity. The pattern of species abundance is shown in Table 2. In general, the number of species in the abundance classes decreased as the abundance increased. Most species (52) were in the first abundance class, making this the

TABLE 1. Time-periods for sampling strategies.

Strategy #	Inclusive time-periods	Extent of sample (h)
1	1–16	2130–0530
2	3–10	2230–0230
3	3–9	2230–0200
4	4–10	2300–0230
5	4–9	2300–0200
6	5–10	2330–0230
7	5–9	2330–0200
8	6–10	2400–0230
9	6–9	2400–0200
10	7–10	0030–0230
11	7–9	0030–0200
12	8–10	0100–0230
13	8–9	0100–0200
14	9–10	0130–0230

commonest class. The apparent paradox is that members of these species were rare with just one moth in each species (see Appendix). The fewest species (3) were in the largest abundance class making this the rarest class but members of these species were abundant (>255 moths in each, see Appendix). Also shown in Table 2 are the frequencies expected from the log series model. The similarity between observed and expected appears close and is confirmed as being a good fit by the deviance *chi*-square value of 8.6. The 5% critical value of the *chi*-square distribution with 7 df is 14.1 indicating that the log series model provides a good description of the data. The overall *alpha* index of diversity was 54.

Single-night samples. The number of moth species and individuals trapped in a single night ranged from a low value of 30 moths in 18 species to a high value of 548 moths in 88 species. Values for *alpha*

TABLE 2. Species abundance frequency distribution of a moth catch in the Acadia Forest Experiment Station compared with expected frequencies from the log series model.

Individuals per species	Number of species		Chi-square
	Observed	Expected	
1	52	53.4	0.04
2–3	47	43.9	0.22
4–7	42	39	0.23
8–15	36	35.4	0.01
16–31	37	31.3	1.04
32–63	22	25.3	0.43
64–127	7	16.9	5.80
128–255	9	7.8	0.18
256–511	2 } 3	1.76 } 1.87	0.68
512+	1	0.11	

Total chi-square = 8.6, $P < 0.5$, $P > 0.1$, df = 7. Last abundance class pooled with previous class to meet requirements of chi-square test (see Methods).

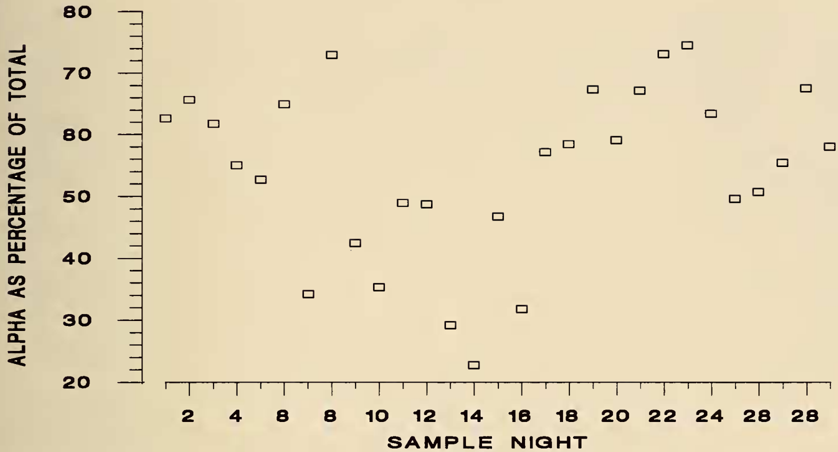

FIG. 1. *Alpha* index of diversity for single-night catches as a percentage of the overall *alpha* based on the total catch.

fluctuated wildly between 12 and 40 with no meaningful trend and never closely approaching the overall *alpha* (Fig. 1). It was apparent that no single-night sample could be used to estimate the index of diversity and thus no pattern of species abundance was determined.

'Replicated' single-time-period samples. For any single 'replicated' time period (consisting of 29, 30-minute samples) the total number of moths trapped ranged between 48 and 627 and the total number of

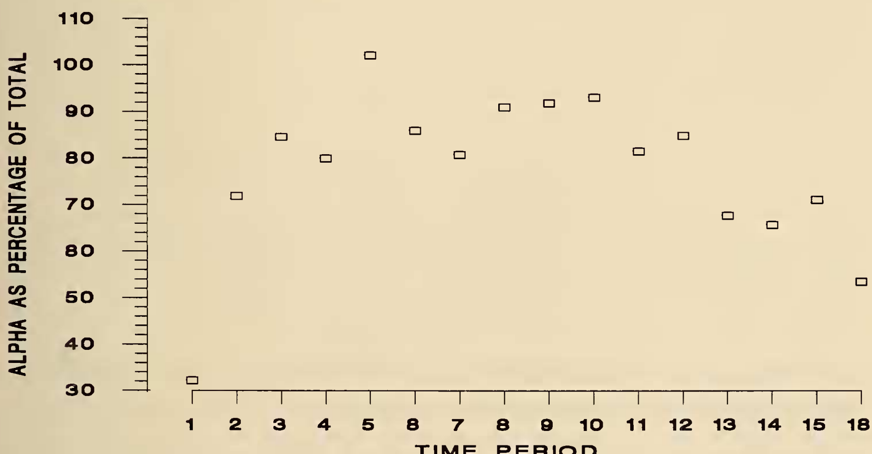

FIG. 2. *Alpha* index of diversity for single-time-period catches, averaged over 29 nights, as a percentage of overall *alpha* based on the total catch. Time periods are sequential 30-minute periods starting at 2130–2200 h and ending at 0500–0530 h.

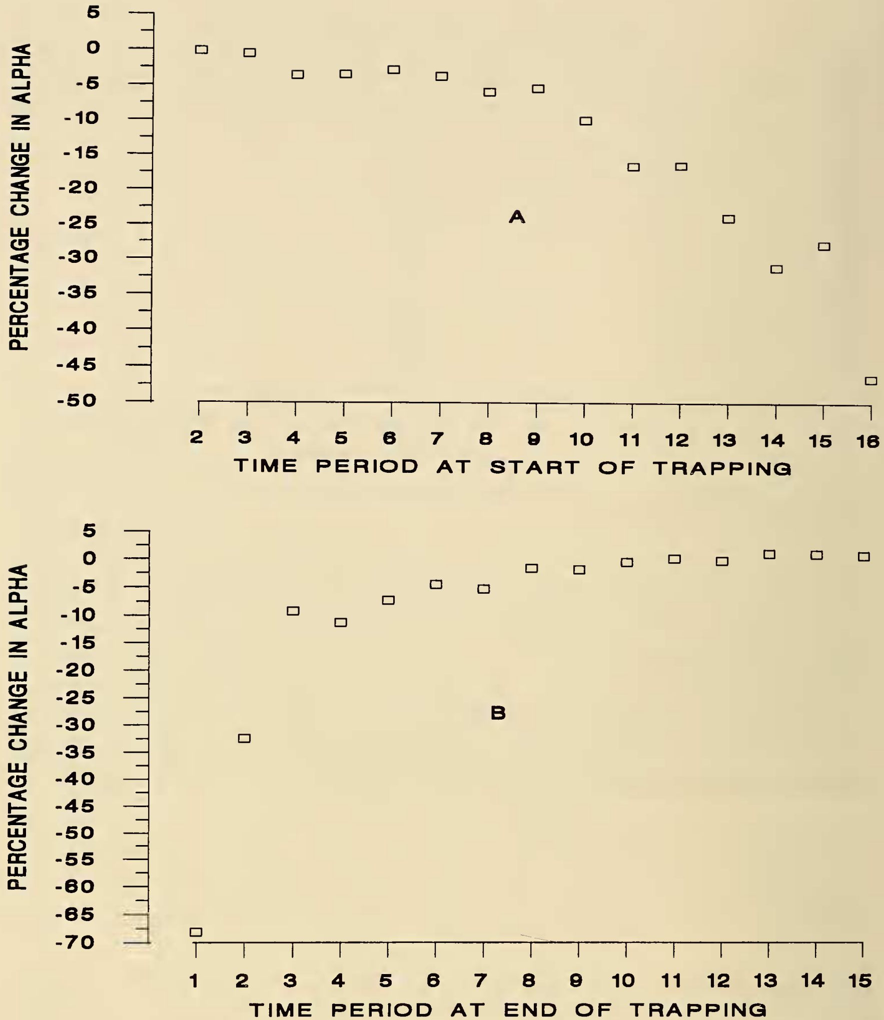

FIG. 3. Percentage change in the *alpha* index of diversity compared to the overall *alpha*: (A) when trapping starts with successively later time periods and ends with time period 16 (0500–0530 h); (B) when trapping starts at time period 1 (2130–2200 h) and end at successively later time periods.

species trapped ranged between 23 and 132. The values for *alpha* for the 'replicated' single-time-period samples started low in the first part of the night, rose rapidly to a maximum during the middle part of the night and then decreased towards dawn (Fig. 2). For time period 5 the value for *alpha* was 102% that of the overall *alpha*. However, this datum was an outlier that did not follow the trend and it was not thought

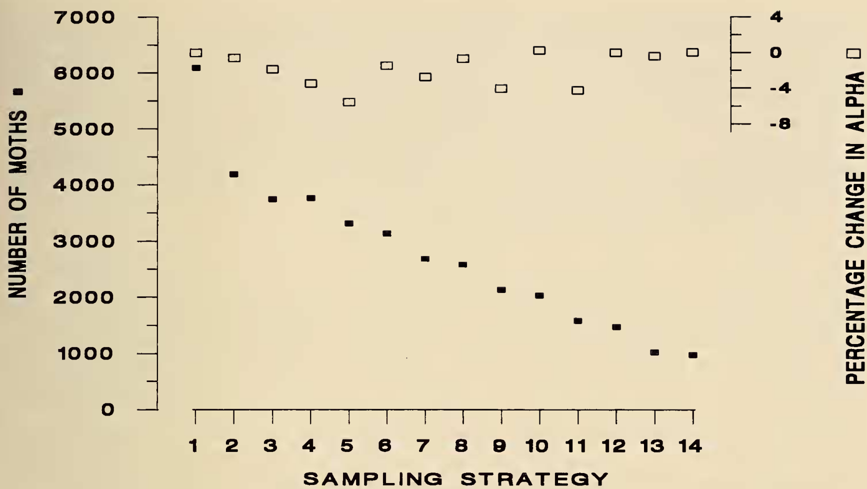

FIG. 4. Percentage change (empty rectangles) in the *alpha* index of diversity relative to the overall *alpha* (strategy 1), and number of moths trapped (solid rectangles) for the various sampling strategies. See methods and Table 1 for explanation of sampling strategies.

prudent to accept this single time period as representative of the overall *alpha*.

Truncated samples—early truncation. Discarding the data in time periods 1 through 6 had no significant effect on *alpha* determined from the remaining data set (Fig. 3A). That is, if the light trap had begun operating at 0030 h, start of period 7, and had run until 0530, *alpha* would have been within 5% of the value obtained by starting the light trap at 2130 h. Also, early truncation of time periods 1 through 6 had no effect on the pattern of species abundance in the remaining data set (time-periods 7–16), *chi*-square = 10.2, 7 df ($P > 0.1$).

Late truncation. Discarding the data in time periods 16 through 9 had no significant effect on *alpha* based on the remaining data set (Fig. 3B). That is, if the light trap had begun operating at 2130 h and had run until 0130 h, the end of period 8, *alpha* would have been within 5% of the overall *alpha*. Also, late truncation had no effect on the pattern of species abundance in the remaining data set (time periods 1–8), *chi*-square = 7.7, 6 df ($P > 0.1$).

Double-ended truncation. Several combinations of early- and late-truncation provided 13 sampling strategies that reduced the sampling period and reduced the number of moths trapped. These strategies (Table 1) had no significant effect on *alpha* and did not compromise the pattern of species abundance. No calculated *chi*-square value, comparison between observed pattern of species abundance and expected pattern from the log series model, was significant (P > 0.05). When the sampling strategies were arranged in a sequence of decreasing sampling periods (Fig. 4), the downward trend in the number of moths trapped and the insignificant effect on *alpha* became obvious. The most cost-effective strategy was a 1-h sample obtained nightly from 0130–0230 h (strategy 14, Fig. 4) that resulted in a total sample of 971 moths in 161 species giving an *alpha* value of 55.

DISCUSSION

The inadequacy of a single-night sample to estimate accurately the *alpha* index of diversity for moths caught during a one-month period was observed by Williams (1943, 1964) in England. Nightly samples during the month of July gave *alpha* values that varied from 42–81% of the overall *alpha* based on the total catch for the whole month, with no evidence of any regular trend (Williams 1964, Table 67). Taylor and Brown (1972) presented data from two traps for nine days in July in Kenya. Single-night *alpha* values ranged from 30.5–80% of the two overall values. Our data showed a similar random pattern with nightly values varying from 22–74% of the overall *alpha* value. Even when Williams (1964, Table 67) calculated diversity on a weekly basis, the average weekly value for *alpha* was only 77% of the monthly value. These data support our conclusion that some sampling effort is required nightly throughout the duration of the calendar dates of interest.

Taylor (1979) commented on the cost-efficiency of sampling insects and the advantages of an attractant trap, such as a light trap, in selecting specific taxa. He also noted that, when used to control pest-species, light traps have as an objective the removal of as large a proportion of the population as possible. However, when used as a monitoring tool, the objective is to affect the population as little as possible compatible with obtaining adequate numbers for analysis. As mentioned in the introduction, large samples have problems associated with the cost of sorting, identifying, counting, and data handling. Reducing sample size by subsampling from a larger sample has drawbacks (Taylor et al. 1979). Taylor and Brown (1972) tried several methods to decrease the size of the moth catch in light traps that included obscuring the light with black paint, changing the source of illumination (different bulb types), and changing the direction of illumination. These methods reduced the size of the catch, but had no effect on the *alpha* index of diversity. They did not examine the effect on the pattern of species abundance.

Our technique of a short-time-period 'replicated' nightly sample to determine the *alpha* index of diversity without changing the pattern of species-abundance is new. Because it results in a relatively small sample, it has the advantage of affecting the moth population much less than a full-night sample. It appears to be of use for determining the moth species diversity of several sites simultaneously which otherwise could not be considered because of processing costs associated with the usually large catches in light traps.

There are no *alpha* index of diversity values from eastern North American forests in similar latitudes with which to compare the *alpha* value obtained in this study. The long-recognized latitudinal and longitudinal gradients in species diversity (Pianka 1966, Smith 1980, see also refs. in Magurran 1988) preclude comparison of the *alpha* value from this study with *alpha* values for moth species diversity in two mid-west American states (Williams 1945) and England (Taylor et al. 1978).

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APPENDIX. Species list with numbers of moths and extreme dates of capture.

Hepialidae		
Korscheltellus gracilis (Grt.)	23–24 July	3
Sesiidae		
Synanthedon acerni (Clem.)	25 June–24 July	11
Cossidae		
Prionoxystus macmurtrei (Guer.)	28 June	1
Tortricidae		
Choristoneura fumiferana (Clem.)	2–29 July	450
Limacodidae		
Tortricidia testacea Pack.	26 June–4 July	4
Tortricidia flexuosa (Grt.)	25 June–29 July	40
Packardia geminata (Pack.)	21 June–29 July	13
Lithacodes fasciola (H.-S.)	26 June–21 July	7
Thyatiridae		
Habrosyne scripta (Gosse)	25 June–18 July	3
Drepanidae		
Drepana arcuata Wlk.	21 June–21 July	20
Drepana bilineata (Pack.)	26 June–29 July	25
Oreta rosea (Wlk.)	10–29 July	5
Geometridae		
Protitame virginalis (Hulst)	21 June–22 July	9
Itame pustularia (Gn.)	14–29 July	183
Itame brunneata (Thunb.)	25 June–17 July	2
Itame anataria (Swett)	17 July	1
Semiothisa aemulataria (Wlk.)	19 July	1
Semiothisa ulsterata (Pears.)	29 June	1
Semiothisa transitaria (Wlk.)	18 July	1
Semiothisa minorata (Pack.)	21 June–29 July	17
Semiothisa bicolorata (F.)	16–19 July	4
Semiothisa bisignata (Wlk.)	29 June–19 July	8
Semiothisa sexmaculata (Pack.)	27 June–24 July	5
Semiothisa signaria dispuncta (Wlk.)	21 June–29 July	724
Semiothisa pinistrobata Fgn.	25 June–25 July	16
Semiothisa orillata (Wlk.)	25–28 June	3
Iridopsis larvaria (Gn.)	21 June–17 July	26
Ectropis crepuscularia (D. & S.)	27 June–29 July	23
Protoboarmia porcelaria (Gn.)	25 June–24 July	5
Melanolophia canadaria (Gn.)	21–26 June	6
Eufidonia convergaria (Wlk.)	25 June–20 July	12
Biston betularia cognataria (Gn.)	27 June–25 July	28
Hypagyrtis piniata (Pack.)	26 June–29 July	193[1]
Lomographa vestaliata (Gn.)	21 June–15 July	30
Cabera erythemaria Gn.	21 June–25 July	41
Cabera variolaria Gn.	21 June–24 July	22
Euchlaena obtusaria (Hbn.)	17–18 July	2
Euchlaena johnsonaria (Fitch)	15–24 July	7
Euchlaena marginaria (Minot)	25 June	1
Euchlaena tigrinaria (Gn.)	4–18 July	2
Euchlaena irraria (B. & McD.)	2 July	4

APPENDIX. Continued.

Xanthotype urticaria Swett	25 June–21 July	5
Pero morrisonaria (Hy. Edw.)	21 June–4 July	13
Nacophora quernaria (J. E. Smith)	26 June–18 July	4
Campaea perlata (Gn.)	21 June–25 July	12
Tacparia atropunctata (Pack.)	27 June	1
Tacparia detersata (Gn.)	21–26 June	8
Homochlodes fritillaria (Gn.)	21–29 June	5
Metanema inatomaria Gn.	21 June–29 July	11
Metanema determinata Wlk.	18–22 July	4
Metarranthis amyrisaria (Wlk.)	21–28 June	3
Metarranthis hypocharia (H.-S.)	25 June	1
Anagoga occiduaria (Wlk.)	21 June	1
Probole amicaria (H.-S.)	21 June–9 July	15
Plagodis serinaria H.-S.	27 June	3
Plagodis phlogosaria (Gn.)	26–29 June	7
Plagodis alcoolaria (Gn.)	21 June–4 July	4
Caripeta divisata Wlk.	25 June–29 July	78
Caripeta piniata (Pack.)	21 June–23 July	9
Caripeta angustiorata Wlk.	17–24 July	22
Besma endropiaria (G. & R.)	21–29 June	6
Sicya macularia (Harr.)	16–25 July	3
Eusarca confusaria Hbn.	16 July	1
Tetracis cachexiata Gn.	21 June–2 July	39
Nematocampa resistaria (H.-S.)	17–29 July	39
Nemoria mimosaria (Gn.)	14–15 July	2
Cyclophora pendulinaria (Gn.)	21 June–25 July	47
Scopula cacuminaria (Morr.)	18 July	1
Scopula limboundata (Haw.)	25 June–24 July	36
Dysstroma citrata (L.)	25–28 June	2
Dysstroma walkerata (Pears.)	21 June–14 July	4
Dysstroma hersiliata (Gn.)	15–29 July	3
Eulithis explanata (Wlk.)	16–29 July	55
Ecliptopera silaceata albolineata (Pack.)	21 June	1
Hydriomena perfracta Swett	21–25 June	2
Hydriomena renunciata (Wlk.)	21 June–29 July	79[2]
Hydria undulata (L.)	14 July	2
Rheumaptera hastata (L.)	16 July	1
Rheumaptera subhastata (Nolcken)	26 June–8 July	2
Mesoleuca ruficillata (Gn.)	25 June	1
Spargania magnoliata Gn.	14 July	1
Perizoma basaliata (Wlk.)	25 July	1
Xanthorhoe abrasaria congregata (Wlk.)	25 June–13 July	8
Xanthorhoe iduata (Gn.)	12 July	1
Xanthorhoe ferrugata (Cl.)	21 June–4 July	3
Xanthorhoe lacustrata (Gn.)	16 July	1
Hydrelia lucata (Gn.)	26 June–18 July	11
Hydrelia inornata (Hulst)	25 June–17 July	9
Eubaphe mendica (Wlk.)	17–20 July	4
Horisme intestinata (Gn.)	29 June	1
Lobophora nivigerata Wlk.	26 June–29 July	63
Lasiocampidae		
Malacosoma disstria Hbn.	9–29 July	136
Malacosoma americanum (F.)	15–25 July	27

APPENDIX. Continued.

Saturniidae		
Dryocampa rubicunda (F.)	21 June–21 July	31
Anisota virginiensis (Drury)	25 June	1
Antheraea polyphemus (Cram.)	21 June–22 July	8
Sphingidae		
Ceratomia undulosa (Wlk.)	21 June	2
Sphinx gordius Cram.	21 June–20 July	9
Lapara bombycoides Wlk.	21 June–24 July	18
Smerinthus jamaicensis (Drury)	21 June–25 July	14
Smerinthus cerisyi Kby.	21–29 June	2
Paonias excaecatus (J. E. Smith)	21 June–23 July	15
Pachysphinx modesta (Harr.)	21 June–23 July	43
Notodontidae		
Clostera apicalis (Wlk.)	21–26 June	2
Nadata gibbosa (J. E. Smith)	21 June–24 July	16
Peridea basitriens (Wlk.)	15–29 July	2
Peridea angulosa (J. E. Smith)	24–25 July	2
Peridea ferruginea (Pack.)	26 June–25 July	150
Pheosia rimosa Pack.	27 June–29 July	8
Odontosia elegans (Stkr.)	17–25 July	2
Notodonta simplaria Graef	15–24 July	7
Gluphisia septentrionis Wlk.	25 June–25 July	54
Furcula cinerea (Wlk.)	29 June–24 July	5
Furcula modesta (Hudson)	16–25 July	11
Symmerista leucitys Franc.	21 June	2
Macrurocampa marthesia (Cram.)	15–25 July	3
Heterocampa umbrata Wlk.	25 June–4 July	11
Heterocampa guttivitta (Wlk.)	29 June	1
Heterocampa biundata Wlk.	21 June–20 July	24
Lochmaeus manteo Doubleday	20–25 July	3
Schizura ipomoeae Doubleday	21 June–24 July	29
Schizura badia (Pack.)	21–27 June	2
Schizura unicornis (J. E. Smith)	15–24 July	10
Schizura leptinoides (Grt.)	25 June–23 July	8
Oligocentria semirufescens (Wlk.)	18–24 July	3
Oligocentra lignicolor (Wlk.)	26 June–29 July	89
Arctiidae		
Eilema bicolor (Grt.)	12–25 July	22
Hypoprepia fucosa Hbn.	4–29 July	54
Haploa lecontei (Guer.-Meneville)	9 July	1
Holomelina laeta (Guer.-Meneville)	29 June–25 July	31
Holomelina aurantiaca (Hbn.)	20 July	1
Holomelina ferruginosa (Wlk.)	11–22 July	7
Pyrrharctia isabella (J. E. Smith)	14 July	1
Spilosoma congrua Wlk.	21 June–5 July	40
Spilosoma virginica (F.)	21 June–20 July	39
Hyphantria cunea (Drury)	21 June–24 July	182
Platarctia parthenos (Harr.)	27 June–17 July	3
Apantesis virguncula (W. Kby.)	27 June–20 July	5
Halysidota tessellaris (J. E. Smith)	13–19 July	2
Lophocampa maculata Harr.	21–29 June	48

APPENDIX. Continued.

Cycnia tenera Hbn.	26 June	1
Ctenucha virginica (Esp.)	10–19 July	4
Lymantriidae		
Dasychira plagiata (Wlk.)	26 June–25 July	69
Leucoma salicis (L.)	4–19 July	7
Noctuidae		
Idia americalis (Gn.)	21 June–29 July	50
Idia aemula Hbn.	14–20 July	11
Idia rotundalis (Wlk.)	21 July	1
Zanclognatha pedipilalis (Gn.)	18–24 July	3
Zanclognatha protumnusalis (Wlk.)	12–22 July	7
Zanclognatha cruralis (Gn.)	21 June–29 July	3
Palthis angulalis (Hbn.)	8–29 July	2
Bomolocha baltimoralis (Gn.)	26 June–20 July	11
Lomanaltes eductalis (Wlk.)	25 June	1
Spargaloma sexpunctata Grt.	21 June–21 July	6
Pangrapta decoralis Hbn.	21 June–20 July	26
Parallelia bistriaris Hbn.	21 June–29 July	5
Catocala sordida Grt.	24–25 July	3
Chrysanympha formosa (Grt.)	9–12 July	12
Autographa precationis (Gn.)	29 June	1
Autographa mappa (G. & R.)	26 June	1
Syngrapha altera (Ottol.)	26 June–21 July	4
Syngrapha octoscripta (Grt.)	14 July	1
Syngrapha epigaea (Grt.)	15–21 July	2
Syngrapha viridisigma (Grt.)	18–24 July	2
Syngrapha alias (Ottol.)	21 June–20 July	22[3]
Syngrapha cryptica Eichlin & Cunningham	24 July	1
Syngrapha rectangula (W. Kby.)	6–25 July	27
Syngrapha microgamma nearctica Fgn.	21 June	1
Plusia venusta Wlk.	17–19 July	2
Baileya ophthalmica (Gn.)	21 June	1
Lithacodia muscosula (Gn.)	21 June–10 July	4
Lithacodia synochitis (G. & R.)	8 July	1
Lithacodia concinnimacula (Gn.)	25 June–4 July	5
Lithacodia carneola (Gn.)	25 June–20 July	21
Leuconycta diphteroides (Gn.)	21 June–19 July	14
Panthea acronyctoides (Wlk.)	21 June–25 July	47
Panthea pallescens McD.	27 June–25 July	29
Charadra deridens (Gn.)	21 June–15 July	21
Raphia frater Grt.	21 June–29 July	152
Acronicta americana (Harr.)	21 June–24 July	18
Acronicta dactylina Grt.	14–25 July	9
Acronicta lepusculina Gn.	29 June–4 July	3
Acronicta innotata Gn.	21 June–25 July	19
Acronicta tritona (Hbn.)	15–19 July	3
Acronicta grisea Wlk.	21 June–24 July	18
Acronicta superans Gn.	15 July	1
Acronicta hasta Gn.	25 June	1
Acronicta fragilis (Gn.)	21 June–25 July	14
Acronicta clarescens Gn.	25 June–25 July	162
Acronicta retardata (Wlk.)	26 June–25 July	49

APPENDIX. Continued.

Acronicta impleta Wlk.	29 June	1
Acronicta noctivaga Grt.	27–28 June	2
Acronicta impressa Wlk.	26 June	1
Acronicta oblinita (J. E. Smith)	26 June–14 July	4
Agriopodes fallax (H.-S.)	25 June–29 July	29
Harrisimemna trisignata (Wlk.)	15–29 July	4
Apamea verbascoides (Gn.)	23 July	1
Agroperina cogitata (Sm.)	10 July	1
Amphipoea velata (Wlk.)	23–25 July	3
Euplexia benesimilis McD.	21 June–25 July	36
Phlogophora iris Gn.	27 June–25 July	3
Chytonix palliatricula (Gn.)	21 June–25 July	78
Dypterygia rozmani Berio	25 June	1
Hyppa xylinoides (Gn.)	18–21 July	4
Nedra ramosula (Gn.)	24 July	1
Callopistria mollissima (Gn.)	25 June–25 July	43
Callopistria cordata (Ljungh)	21 June–29 July	162
Proxenus miranda (Grt.)	4 July	1
Elaphria versicolor (Grt.)	21 June–13 July	51
Elaphria festivoides (Gn.)	25 June–20 July	130
Apharetra purpurea McD.	15–29 July	24
Oncocnemis riparia Morr.	14 July	1
Polia nimbosa (Gn.)	24–25 July	4
Polia imbrifera (Gn.)	13–25 July	6
Polia purpurissata (Grt.)	24–25 July	3
Polia detracta (Wlk.)	4–20 July	8
Polia goodelli (Grt.)	16 July	1
Polia latex (Gn.)	21 June–10 July	18
Melanchra adjuncta (Gn.)	21 June–24 July	25
Melanchra assimilis (Morr.)	26 June–25 July	12
Lacanobia subjuncta (G. & R.)	24 July	1
Lacanobia grandis (Gn.)	21–28 June	14
Lacanobia lutra (Gn.)	21 June–25 July	89
Lacanobia rugosa (Morr.)	27 June–16 July	2
Lacanobia legitima (Grt.)	29 June–25 July	10
Papestra biren (Goeze)	27 June	1
Lacinipolia lustralis (Grt.)	27 June–24 July	17
Lacinipolia anguina (Grt.)	27 June	1
Lacinipolia renigera (Steph.)	29 July	1
Lacinipolia lorea (Gn.)	26 June–18 July	7
Lacinipolia olivacea (Morr.)	29 July	1
Leucania multilinea Wlk.	9–25 July	8
Leucania insueta Gn.	26 June–21 July	24
Leucania inermis (Fbs.)	9–16 July	4
Leucania pseudargyria Gn.	4 July	1
Homorthodes furfurata (Grt.)	2–24 July	77
Orthodes crenulata (Butler)	25 June–25 July	18
Orthodes cynica Gn.	21 June–29 July	268
Euxoa divergens (Wlk.)	5–19 July	2
Ochropleura plecta (L.)	21 June–25 July	29
Diarsia jucunda (Wlk.)	13–29 July	23
Eurois astricta Morr.	24–29 July	5
Xestia dolosa Franc.	15–29 July	10[4]
Xestia oblata (Morr.)	8–20 July	3

APPENDIX. Continued.

Anomogyna elimata (Gn.)	25–29 July	2
Anomogyna badicollis (Grt.)	24–29 July	5
Anomogyna youngii (Sm.)	15 July	1
Aplectoides condita (Gn.)	21 June–4 July	25
Anaplectoides prasina (D. & S.)	8–29 July	8
Anaplectoides pressus (Grt.)	15–25 July	4
Eueretagrotis perattenta (Grt.)	17–25 July	5
Eueretagrotis attenta (Grt.)	8–29 July	60
Heptagrotis phyllophora (Grt.)	27 June–25 July	39
Cryptocala acadiensis (Bethune)	22–24 July	4
Noctua pronuba L.	23–25 July	3

[1] Identification uncertain, may include or consist entirely of *Hypagyrtis unipunctata* (Haworth) (Geometridae).
[2] Includes *Hydriomena divisaria* (Walker) (Geometridae).
[3] Includes *Syngrapha abstrusa* Eichlin & Cunningham (Noctuidae).
[4] Identification uncertain, may include or consist entirely of *Xestia adela* Franclemont (Noctuidae).